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Research progress is reported on (1) biotic associates of free-living stages of chigger mites, (2) extraction efficiencies for different methods, (3) food preferences of adult Eutrombicula, and (4) vertebrate host associations for the chigger fauna. Adult Eutrombicula densities were in the range of 1-10 per m^2, based on flotation of soil samples. Flotation in salt solution or tap water yielded 90% recovery, but so did 5-day tullgren extractions. The latter are for less labor-intensive. Eutrombicula adults

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were offered eggs of eight different soil collembolan species. Adults showed a high preference for eggs of only three of the species. Vertebrate host associations in Georgia were dominated by Eutrombicula species. Infestation rates were higher on the Coastal Plain than on the Piedmont, and higher for reptiles than for birds or mammals.

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ANNUAL TECHNICAL REPORT NUMBER 2

Ecological Requirements of Chigger Mites

by

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INTRODUCTION

Chigger mites are vectors of disease in some parts of the world and are pests of major importance. Bites of some chigger species produce a mild to severe dermatitis in man and some animals, causing manpower and economic losses. Chiggers, the larval stage of trombiculid mites, are vectors of scrub typhus, a rickettsial disease, in parts of the asiatic-pacific area (Traub and Wisseman 1974). In Vietnam, scrub typhus was responsible for more "fevers of unknown origin" in American troops than any other tropical disease except malaria (Nadchatram and Dohany 1974). In the United States turkey chiggers (*Neoschoengastia americana*) are responsible for economic losses in turkey production (Kunz 1969). Chiggers causing dermatitis in humans (pest chiggers, *Eutrombicula* spp.) affect use patterns in recreational areas. Military training exercises in the southeastern United States may be severely impacted by both chiggers and ticks. In sensitive individuals, dermatitis caused by chigger infestations may result in hospitalization. As a result of their importance to man, chiggers have been the subject of an extensive taxonomic literature devoted to the parasitic larval stage. Little is known of the free-living nymphal and adult stages, except that they are non-parasitic predators living in soil or similar habitats. The last major taxonomic work on the postlarval stages of trombiculids (Crossley 1960) was published 20 years ago.

The general life history of trombiculids is well known (Loomis 1956). Eggs develop through a deutoval stage and hatch into the hexapod larval stage, the "chigger." Hosts of the parasitic chigger stage are vertebrates:

amphibians, reptiles, birds and mammals. Host specificity varies among the hundreds of known chigger species, from a highly specific to an extremely broad range of vertebrate hosts. The taxonomy of the group is based in large part upon the larval stage. Engorged larvae drop from the host and pass through protonymph (quiescent), deutonymph (active), tritonymph (quiescent) and adult (active, bisexual) stages. The active forms (deutonymph and adult) are predators on small arthropods and their eggs. These active postlarval stages are little known. Most have not been collected, reared or described. Most knowledge of postlarval stages has been gained from laboratory rearings, starting with engorged larvae obtained from vertebrate hosts. To rear trombiculids to the adult stage, a suitable food for the predaceous deutonymphs must be found. A variety of arthropod prey species have been successful in laboratory rearings (Lipovsky 1954), although the food items are probably not the normal ones. Success has been achieved with foods which trombiculids might not encounter under natural conditions. For example, Nadchatram (1968) used culicine mosquito eggs as a standard laboratory food for deutonymphs and adult trombiculids. American workers have used, among other things, eggs or immatures of the collembola Sinella curviseta, a species easily bred in the laboratory (Lipovsky 1951). Using these foods, trombiculids have been reared in the laboratory and a few species induced to reproduce (Nadchatram 1968). The establishment of laboratory culture of chiggers has been hampered by (1) lack of a suitable food for postlarvae and (2) lack of knowledge of abiotic requirements of postlarvae.

Only a few trombiculid species have been collected as postlarval

stages (Crossley 1960), but conclusions about the postlarval habitat may be drawn from host associations of the chiggers themselves. Identification of the postlarval habitat is based upon the range of vertebrate hosts on which the chigger species occurs, and knowledge of the ecology of each host (Loomis 1956, Crossley 1960). Some highly host-specific species are probably nest inhabitants as postlarvae. For those chiggers which occur on a variety of vertebrate hosts, frequency analysis may suggest which hosts are most likely to encounter chiggers. Postlarval habitats may then be inferred from knowledge of the habitats of the hosts. The most extensive analysis of this type was performed by Nadchatram (1970) for Malayan chiggers. He recognized seven ecological groups of trombiculids, based on occurrence in local habitats, host associations and color of unfed larvae. Adult trombiculids were then located in the field, in two of the suggested field habitats for the seven groups.

Research reported here was undertaken to address questions about the ecology of freeliving stages of chigger mites, by identifying their habitats, the biotic and abiotic environmental factors which predispose certain areas to the presence of certain chigger species, and the general ecological requirements of the adult stages. Also necessary is knowledge of the host associations, as well as suitable laboratory rearing methods, so that biotic and abiotic factors can be evaluated experimentally.

Our first year's research was devoted to initiation of both laboratory and field projects, with emphasis on field collection of hosts, detection of postlarval habitats, and improvements in collection methods for both larvae and adults. In addition, development of mass rearing

methods was a major undertaking.

In this, the second annual report we emphasize progress in four areas: (1) identification of postlarval (adult) environments and their biotic associates, (2) extraction methods for postlarvae, (3) food preferences of adult chiggers and (4) analysis of trombiculid-vertebrate host relations. These reports should be considered preliminary, since interpretations are subject to revision. All research projects are considered as on-going for the next year.

Beginning with our second year, we have focused our attention on pest chiggers of the genus *Eutrombicula*. These species are medically important, and are the most abundant ones found by sampling plates and on vertebrate hosts. *Eutrombicula* occurs in a variety of habitats, and may be considered to be a generalist. Since our principal research focus is on the post-larval habitat, working with a generally distributed group of species may prove to be a disadvantage. However, the results should have broad application. Had we concentrated on more specialized species of chiggers (amphibian or bat associates), any "key" biotic factors identified would probably be highly specific.

PROGRESS IN RESEARCH PROJECTS

Postlarval environments and biotic associates

We have completed one years field sampling for postlarval stages (nymphs and adults) of *Eutrombicula alfreddugesi*, using flotation methods for extraction. Soil samples have been taken from selected habitats (non random), from an unbiased series (random), and from transects (regular) established in areas where larvae (chiggers) were present. At

the postlarval sampling sites, we have sampled soil arthropod associates of chiggers using high-efficency extraction of soil cores (Merchant and Crossley 1970). Data from these collections are currently being analyzed, and are not ready for summarizing and reporting. For purposes of this Annual Report, we give below the current status of our research findings.

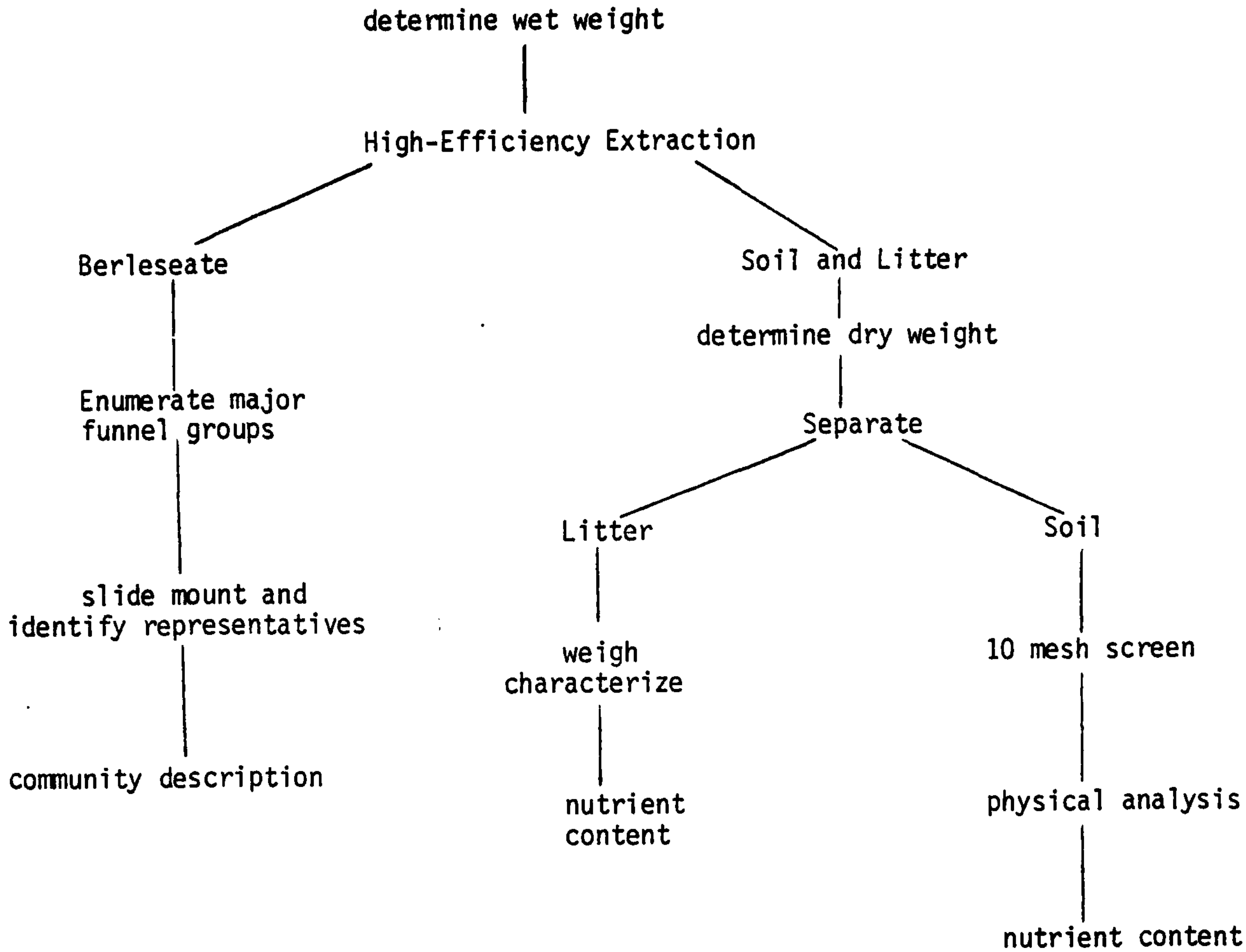
Postlarval habitat - -. We have been moderately successful in locating adult Eutrombicula in soils at sites where larvae occurred on chigger sampling plates. Last year we suggested that decomposing pine logs (*Pinus taedia*, *P. echinata*) were postlarval habitats. Further collections now indicate that soil itself is the habitat, and that the presence of underground organic debris is also important. For example, adult *Eutrombicula* are associated with decomposing pine stumps and are specifically found in mineral soil which is in contact with subterranean decomposing bark and wood. Similarly, adult *Eutrombicula* were found on coastal dunes associated with buried wood and leaf litter. The presence of vertebrate hosts for the larval stages is also an important correlate. The pine stumps mentioned above are used by lizards (*Eumeces laticeps* - *Sceloporus undulatus*) as "roosts." Pine logs, and even boulders, where unengorged larvae occur are those used by the hosts. Our current perception, then, is that nymphs and adults live in soil habitats associated with buried organic litter. Eggs are laid there. Upon eclosion, larvae climb vertically to the roosts of hosts. Engorged larvae fall back to the soil surface.

Postlarval numbers. - - Our sampling suggests that *Eutrombicula* densities are lower than expected -- in the range of 1-10 per m^2. This

density is an order of magnitude below that reported by Jenkins (1947). Our results are based primarily on flotation of soil samples, and efficiencies for this method are satisfactorily high (see Extraction methods for adult *Eutrombicula* below). We will continue to measure postlarval densities.

Biotic associates of postlarvae. - - This data set is based on high-efficiency extraction of cores taken in conjunction with samples of chiggers. The analytical scheme for these soil cores (Table 1) allows for estimation of physico-chemical properties of the soil as well as description of the arthropod community. In addition, a seasonal photographic record is maintained for each sampling site. Currently, two transects, each 24 m long, are being followed in two different habitat types. Permanent stakes mark the location of sampling points along each transect. During the summer, densities of unengorged larvae were recorded biweekly at each stake. We are using the presence of unengorged larvae as evidence that the soil habitat at that point is capable of supporting adults. During the summer, those sample stations which had unengorged larvae continued to produce them throughout the season. Sampling points which had not revealed chiggers by July did not develop infestations later. Thus, it appears that the localized presence or absence of unengorged larvae was a repeatable phenomenon. The presence of these larval clusters may indicate that eggs were laid at that site, or that larvae were clustering there in response to some environmental cue. Larvae are capable of some motion, but directed movements have only been reported for transient environmental stimuli (temperature, humidity,

Table 1. Analytical scheme for soil core samples



CO_2,light) (Jenkins 1947). Such transient stimuli would not account for the presence of unengorged larvae at a particular spot on the soil surface for weeks at a time. It will be interesting to see whether the same sampling spots become infected next summer.

Extraction Methods for Postlarval Trombiculids

The low numbers of adult *Eutrombicula* recovered from samples caused us to re-examine the efficiency of our extraction methods. We compared the Ladell apparatus (Lawson and Merritt 1979) with floatation in epsom salts and tap water. Also, we compared Tullgren funnel extraction (with and without lights) to floatation. Satisfactory recoveries were obtained with all methods (including the flotation we have been using) except the Ladell apparatus. In the future we will use Tullgren funnels since that method is the least labor-intensive.

All of the extraction techniques evaluated in this investigation began by placing mites and soil into a series of plastic bags. Each bag contained 550 grams of autoclaved soil and surface litter. Five postlarvae were carefully placed into each bag, using a camel hair brush. A total of five replicate bags of soil, litter, and postlarvae were used for each extraction method. Five hundred fifty grams was the mean sample size examined for adult *Eutrombicula* during the past year. Five postlarvae per bag were chosen because five is the most postlarvae we have collected from any single sample, and owing to the low availability of postlarval chiggers for use in these experiments, optimization between sample size and replicate number was essential. Postlarval chiggers were left in the bags to disperse through the soil for one hour prior to extraction.

For flotation extraction, comparisons were first made between the efficiency of magnesium sulfate (epsom salt) solution (100 gms per liter, specific gravity 1.06) and tap water. Two liters of epsom salt solution or tap water were poured into a square plastic basin. A bag containing the soil, litter and postlarvae was then emptied into the basin. After an initial scan of the surface, the contents of the basin were stirred vigorously. Again the surface was scanned, with any postlarval chiggers floating on the surface being removed with a camel hair brush. This process was repeated until all the postlarvae were recovered, with a time limit of 20 minutes. This same procedure was repeated for all five replicates, for both the tap water and epsom salt solution.

The efficiency of the Laddell apparatus for extracting postlarval chiggers reported last year was evaluated. Five bags of soil and litter, each containing five postlarvae, were extracted.

The efficiency of funnel extractors was evaluated using 10 funnels. Tullgren funnels were 30 cm dia, with regulated heat sources provided by 40 watt bulbs. Each funnel extracted a sample consisting of 550 grams of soil, litter and 5 postlarvae. Five of the funnels utilized light sources, while five other funnels were left without lights. Temperature probes were placed in the center of one soil sample in both the light and dark funnels. Extracted postlarvae were collected onto the surface of water filled vials underneath the funnels. The vials and temperatures were checked every four hours. Extraction was terminated after one week.

Table 2 presents a summary of the results of the extraction method comparisons. Excellent recoveries (90% or better) were demonstrated for

Table 2. Comparison of Extraction Efficiencies For Postlarval Eutrombicula. Each mean based on 5 replicates, with 5 postlarvae per replicate.

Method	mean recovery (of 5)	S.E.	percent recovered (efficiency)	mean number alive (of 5)	percent alive
Floatation					
Epsom salts	4.6	.134	92	3.6	78
Tap water	4.8	.089	96	4.6	96
Ladell	2.8*	.313	56	2.6	93
Tullgren Funnel					
with light	4.8	.089	96	-**	-
without light	4.2*	.313	84	-	-

* recoveries significantly lower ($P < .05$).

** all mites recovered were alive.

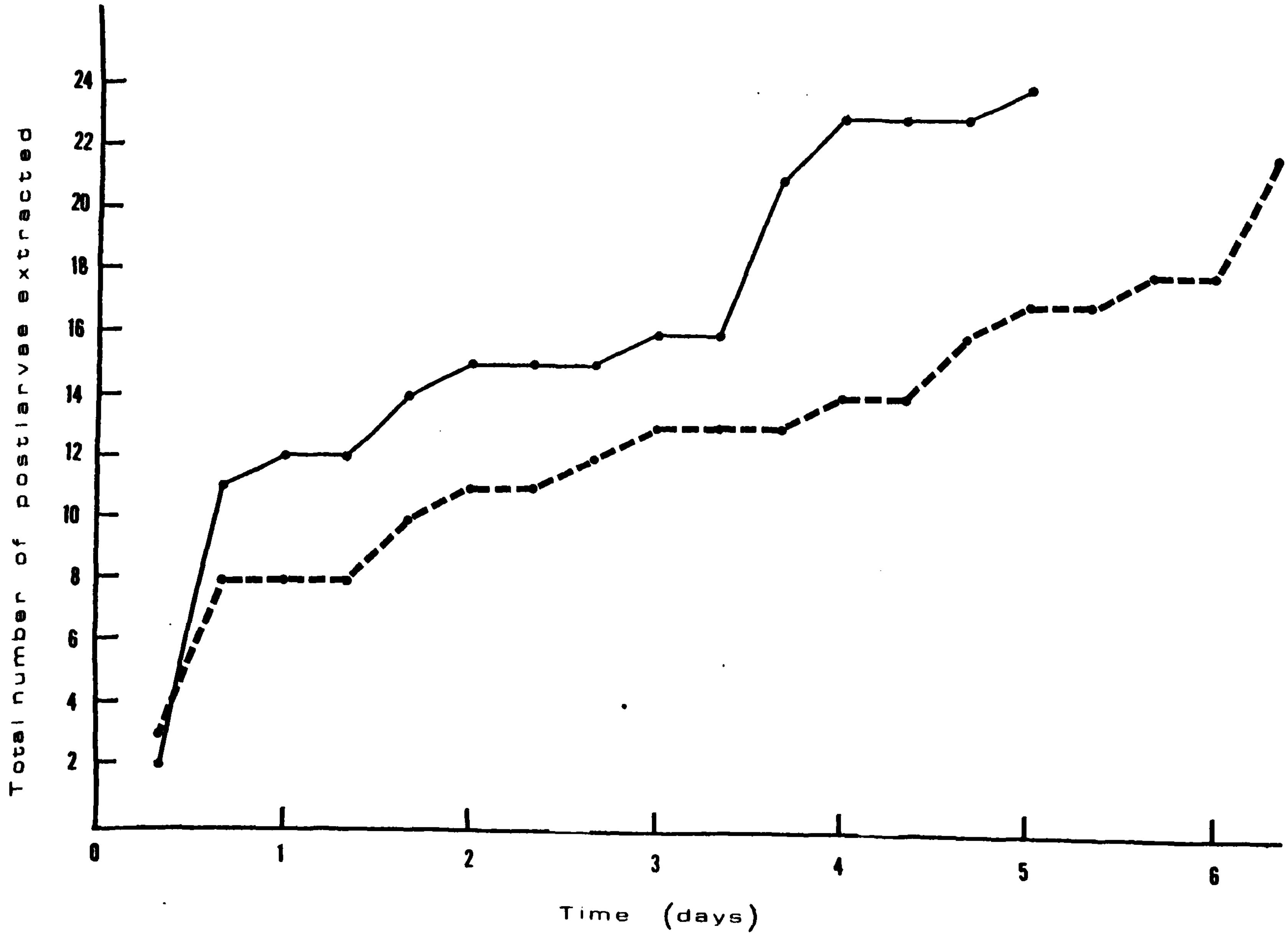
floatation in either epsom salts or tap water, and for lighted Tullgren funnel extraction. The Ladell apparatus and the unlighted Tullgren funnels gave less satisfactory responses. Because Tullgren funnels require less labor, we will use them for extractions of future samples.

The high efficiency of the Tullgren funnels was unexpected. Traditionally, acarologists believe that Trombiculids do not respond well to Tullgren or Berlese funnels. Our inclusion of the unlighted Tullgren funnels in the experimental design was intended to provide a more gradual dessication and to negate any positive phototaxis. The unlighted funnels were cooler (mean temperature 26.5^{o}) than the lighted ones (mean temperature 31.2^{o}) or the ambient temperature in the room (28.6^{o}). Time-phased collections (Figure 1) indicate that extraction efficiency was nearly linear over a 5-day period. Tullgren funnels with lights had an extraction half-time (time required to extract 50% of the mites in a sample) of 28.4 hrs ($r^2 = .916$, $n = 10$). Unlighted funnels had an extraction half-time of 66 hrs ($r^2 = .879$, $n = 12$). Thus, a 6-day extraction period on lighted funnels should yield 95% recovery of trombiculid mites in a 550 gram soil sample.

Food Preferences of Adult Trombiculids

The objective of this research was to evaluate food preferences of adult *Eutrombicula splendens* for eggs of a variety of collembolan species. Collembolans are common biotic associates of postlarval trombiculids, and their eggs or other instors may be important food sources in natural habitats. Also, collembolan eggs (especially *Sinella curviseta*) have been used as food sources for laboratory rearings of *Eutrombicula* and

Figure 1. Cumulative recovery rates for twenty-five adult Eutrombicula splendens placed in 500 g soil and extracted in Tullgren funnels with (.—.) or without (.---.) lights as heat sources.


Total number of postlarvae extracted
24
22
20
18
16
14
12
10
8
6
4
2
0
1
2
3
4
5
6
Time (days)

other trombiculid genera (Lipovsky 1951, 1954; Everett, Price and Kunz 1973; Cunningham, Kunz and Price 1977; Simonova 1977; Huber 1978). Other food materials used for rearing trombiculids (mosquito eggs, dissected ovarian materials) have been relatively convenient but unnatural, since trombiculids do not encounter them in natural habitats (Jaywickreme and Niles 1946, 1947; Lipovsky 1954; Kulkarni and Mahaden 1973; Jenkins 1947). Also, the use of unnatural foods greatly increases the labor necessary for maintenance of cultures. Collembolans, on the other hand, can be maintained in colonies with the trombiculids. Collembolans have a high intrinsic rate of increase (Gist, Crossley and Merchant 1974) and can provide a constant food source for the predaceous postlarval trombiculids.

Materials and methods. - - Field collected Eutrombicula splendens adults from the south coastal region of Georgia were each placed in separate plastic cells with charcoal-plaster substrates. Three holes drilled in the bottom of the plastic cells before the substrate was added, providing a means of water absorbtion and drainage. This arrangement maintained a constant high humidity with a minimum of disturbance to the mites. Cells containing mites were kept at a constant 28^{o} C in an environmental chamber with a 14/10 day/night cycle, for 6 days prior to and during experiments. Each adult mite was randomly assigned to one of 8 treatment groups. Each treatment group was composed of 8 adults. Each treatment consisted of provisioning each cell with 30 freshly laid eggs of one of the following collembolan species: Hypogastura armata, Onychiurus encarpatus, Onychiurus folsomi, Proistoma minuta, Pseudosinella sexoculata, Pseudosinella violenta,

Sinella curviseta and Tullbergia krausbueri. The number of eggs consumed was determined 24 hours later. Data analyses utilized one-way analysis of variance and Duncan's new multiple range test (Ott 1977).

Results. - - Analysis of variance showed a strong difference in the feeding responses of adult Eutrombicula to collembolan eggs of different species ($F = 39.20$, $P < 0.001$). S. curviseta eggs were the most acceptable food source with an average of 17.5 eggs eaten per adult per day (Table 3). This consumption was significantly greater than for eggs of any of the other species. The eggs of both T. krausbueri ($\bar{X} = 11.25$) and P. violenta ($\bar{X} = 10.75$) were equally preyed upon, although to a lesser extent than those of S. curviseta. Both of these experimental groups had relatively large standard errors (1.8588 and 1.8874) (Table 1) when compared with the other treatments, indicating wide differences in individual mite response. Individual responses to T. krausbueri ranged from 4 to 20 eggs consumed and for P. violenta from 4 to 18. O. folsomi eggs were consumed, but at a much lower rate (3.88 eggs per adult per day) than those previously mentioned. Their low consumption rate places them in a low preference category even though they were eaten at a significantly greater rate than the remaining species. The last 4 species, P. sexoculata, H. armata, P. minuta and O. encarpatus belong to a statistical population which was not acceptable to the postlarval trombiculids as a food source. The 1 or 2 eggs destroyed per experimental unit may be considered the result of experimental handling or possibly "tasting" by the mite. The comparatively low consumption rates (0.25-1.1 eggs per adult per day) indicate that eggs of these collembolan species are inadequate or

Table 3. Suitability of different species of Collembolan eggs as food for adult Eutrombicula splendens. $\bar{X}$ = eggs consumed per adult per day, S.E. = standard error. Variates are means of eight replicates.

	Sinella curviseta	Zulbergia krausbueri	Pseudosinella violenta	Onychiurus folsomi	Pseudosinella sexoculata	Hypogastura armata	Proistomia minuta	Omychiurus encarpatus
$\bar{X}$	17.50	11.25	10.75	3.88	1.13	0.75	0.38	0.25
S.E.	.8452	1.8874	1.8588	.8544	.4407	.3134	.1830	.1637
Statistical population*	1	2	2	3	4	4	4	4

* Collembolan species sharing the same number are not significantly different (P >0.05).

unacceptable as food sources.

The differences in food preference exhibited by the chiggers demonstrates that the mites are able to discriminate between potential food supplies in their natural environment, but the mechanism for discrimination is not obvious. It does not appear to be based on the size, texture, or rigidity of the egg shell. Under the dissecting scope, the texture of all eggs is uniformly amorphic. The shells seem to be equally fragile and can be broken by the touch of a single hair from a sable brush. _S. curviseta_, _P. sexoculata_, _P. violenta_, _H. armata_, _O. folsomi_ and _O. encarpatus_ all lay eggs of a very similar size. _P. minuta_ and _T. krausbueri_ lay smaller eggs: _T. krausbueri_ is eaten and _P. minuta_ is not. Thus size alone is not a factor in food selection. Lipovsky (1954) stated that only entomobryids, in particular, _S. curviseta_, are suitable for culturing with chiggers. Our results show that high preference collembolan species may not be grouped according to Family. _P. sexoculata_, _P. violenta_ and _S. curviseta_ are all members of the Family Entomobryidae, but _P. sexoculata_ is not eaten al all while both the others are heavily consumed. The family Brachystomellidae contains _O. encarpatus_, _O. folsomi_ and _T. krausbueri_, yet only _T. krausbueri_ was preferred. The variability observed in the feeding responses to _T. krausbueri_ and _P. violenta_ is not readily explainable. The chiggers were randomly assigned to each treatment and thus variability due to satiety, physiological state and previous experience should be evenly distributed among all treatment groups. Some variability may exist within the eggs of both these species. A genetically induced chemical difference or a biotic contaminant may render the egg unpalatable

to the chiggers.

Analysis of trombiculid-vertebrate host relationships

Distribution of trombiculid mites is generally correlated with distribution of potential vertebrate hosts (Jenkins 1947, Srivastva and Wattal 1973). Most trombiculids exhibit little host specificity (Buckner and Gleason 1974, Loomis 1956). Of those that do, some may be host-specific due to limited availability of hosts in the postlarval habitat, and others may not find suitable attachment sites except on certain hosts (Loomis 1956, Wharton 1957, Nutting 1968). The most abundant and widespread chiggers are host generalists and thus tend to be pest chiggers of man (Jenkins 1947, Crossley 1960, Crossley and Proctor 1971). Important examples are found in the genus *Leptotrombidium*, which is a vector of the rickettsial disease scrub typhus in southeast Asia (Traub and Wisseman 1974, Hubert and Baker 1963, Nadchatram 1970). In the southern United States, pest chiggers of the genus *Eutrombicula* are universally distributed and sometimes medically important (Jenkins 1947, Crossley and Proctor 1971). Analysis of distribution of chiggers within and among hosts is a promising way to approach questions of abundance, species composition, and distribution of trombiculid mites in relation to season, habitat, and geography. In this section, we report on data collected in 1980 and 1981 on host-parasite relationships of chigger mites in Georgia.

Vertebrate hosts of trombiculid mites were taken by snap trap, live trap, pellet gun, shotgun, and hand collection. Collecting effort was concentrated in the Piedmont region, but over 125 sites were sampled across the state of Georgia. The sites were assigned to geographical

region following the terminology of Golley (1962), modified to consolidate his mountain categories. Assignment of sites to habitat group is explained in the following section. Chigger mites were recovered from hosts by methods reported in Wicht and Crossley (1982) and in last year's Annual Report. Generally, reptiles were placed live over water and engorged trombiculid larvae recovered from the water's surface. Mammals and birds were killed and washed in a solution of detergent and tap water which was decanted into a separatory funnel for recovery of live, undamaged larvae. Larvae were mounted on permanent slides for identification. In the case of heavily infested hosts, a sample of the chiggers was mounted and the excess used to establish and maintain cultures.

Over a two year period, we have recovered more than 5000 larval chiggers from 850 vertebrate hosts (Table 4). Most vertebrates were taken on the Piedmont, and we have classified sample sites on the Piedmont into three habitat types: mature forest, disturbed forest, and disturbed open systems. Mature forests are those which have grown through successional stages to a "climax" community with only widely scattered large pines. Most of our mature forest sites were in riverbottom hardwood forest, dominated by *Acer negundo*, *Betula nigra* and *Liquidambar styraciflua* with DBH greater than 35 centimeters. Ground cover in this habitat is sparse, and consists primarily of young trees, ferns and privet (*Ligustrum* sp.). Our disturbed forest sites comprise a heterogeneous assemblage of habitats encompassing successional sera from late stage old fields to mixed pine and hardwood forest. Monotypic pine stands are included in this category, and dominant trees had DBH less than 35 cm. A shrub understory

Table 4. Host-parasite data for all species of larval trombiculids (chiggers) found on all species of vertebrate hosts examined, 1980-1981

	number of hosts examined	number of hosts infested	percent of hosts infested	total number of chiggers removed	number of chiggers per infested host
Georgia (Total)	850	114	13.4	5024	44.1
Coastal Plain	22	17	77.3	961	56.5
Piedmont	828	97	11.7	4063	41.9
mature forest	36	0	-	-	-
disturbed forest	65	27	41.5	2341	86.7
disturbed open	727	69	9.5	1713	24.8

was often present, commonly dominated by Celtis, Vaccinium, Oxalodendron, Cornus, and Liriodendron. Ground cover ranged from sparse to dense, and was usually a mixture of grasses, forbs, Smilax and Rubus. Disturbed open sites were old fields in various stages of succession, usually with scattered young pines and pioneer hardwoods, and often near buildings, roadways, powerlines, or other foci of human activity.

Table 4 displays chigger infestation data in relation to physiographic region and habitat of capture of all potential hosts. The infestation rate for Georgia as a whole was low (13.4%), as was that for the Piedmont (11.7%). The Coastal Plain had a much higher infestation rate (77.3%), although the number of chiggers per infested host was only slightly higher. Most of our specimens were taken on the Piedmont in disturbed open habitats. This is partly due to differential sampling; that is, these habitat types are common and accessible and so received disproportionate trapping effort. In addition, review of field notes suggests that catch per trap night was lower in both forest situations. Despite reduced effort and catch efficiency, disturbed forest situations were far more productive of chiggers than either mature forest or disturbed open habitats. Both frequency and severity of infestation of all hosts were highest in disturbed forests. The extremely high severity value (130 chiggers per host) is at least partly due to a single cottontail rabbit (Sylvilagus floridanus) from which we recovered 1598 larval chiggers -- more than four times as many as from any other single host. Cottontails in general, although poorly represented in our collections, had very high rates and severities of infestation. Lagomorphs could be foci of chigger larvae in

their habitat, much as Mus and Rattus are in settled regions of India (Srivastva and Wattal 1973).

Tables 5-9 show host-parasite data for major taxa in a format similar to that of Table 4. We examined only 64 birds (mainly starlings, Sternus vulgaris) and consider our data too weak to support generalization (Table 5). In view of published information (Loomis 1956, Crossley and Proctor 1971, Wrenn 1974) we expect birds to be infested at low rates and severities, although nidicolous fauna and species composition of avian trombiculid parasites may prove to be different. Small mammals (Table 6) comprise the bulk of specimens which we examined for chiggers. Again, the rate of infestation of mammals taken on the Coastal Plain (42.9%) was greater than that of animals from the state as a whole (9.7%) or from the Piedmont (9.3%). Severity of infestation was 41.3 chiggers per host in all physiographic regions, although variance was reduced on animals from the Coastal Plain (Table 6). Thus, chiggers were distributed more evenly within hosts as well as among hosts on the Coastal Plain. On the Piedmont, disturbed forest habitats had highest rates and severities of infestation. Two species, Peromyscus leucopus (Table 7) and Sigmodon hispidus (Table 8) were taken in sufficient numbers for detailed analysis. The cotton rat, S. hispidus, was taken only in disturbed habitats and had an infestation rate (7.6%) and severity (10.0 chiggers per host) that were greater than those recorded for the white-footed mouse, P. leucopus (3.8% infestation, 2.0 chiggers per host). P. leucopus was lightly parasitized even in disturbed open habitats in which S. hispidus was infested at twice the rate and five times the severity of P. leucopus. We believe that

Table 5. Host-parasite data for all species of larval trombiculids (chiggers) found on all species of birds examined, 1980-1981

	number of hosts examined	number of hosts infested	percent of hosts infested	total number of chiggers removed	number of chiggers per infested host
Georgia	64	3	4.7	3	1.0
Coastal Plain	1	1	100.0	1	1.0
Piedmont	63	2	3.2	2	1.0
mature forest	5	0	-	-	-
disturbed forest	4	0	-	-	-
disturbed open	54	2	3.7	2	1.0

Table 6. Host-parasite data for all species of larval trombiculids found on all species of mammals examined, 1980-1981

	number of hosts examined	number of hosts infested	percent of hosts infested	total number of chiggers removed	number of chiggers per infested host
Georgia	703	68	9.7	2811	41.3
Coastal Plain	7	3	42.9	124	41.3
Piedmont	696	65	9.3	2687	41.3
mature forest	30	0	-	-	-
disturbed forest	35	8	22.9	1986	248.2
disturbed open	631	56	8.9	692	12.4

Table 7. Host-parasite data for _Peromyscus leucopus_, 1980-1981

	number of hosts examined	number of hosts infested	percent of hosts infested	total number of chiggers removed	number of chiggers per infested host
Georgia	158	6	3.8	12	2
Coastal Plain	0	-	-	-	-
Piedmont	158	6	3.8	12	2
mature forest	10	0	-	-	-
disturbed forest	26	0	-	-	-
disturbed open	122	6	4.9	12	2.0

Table 8. Host-parasite data for Sigmodon hispidus, 1980-1981

	number of hosts examined	number of hosts infested	percent of hosts infested	total number of chiggers removed	number of chiggers per infested host
Georgia	409	31	7.6	310	10
Coastal Plain	0	-	-	-	-
Piedmont	409	31	7.6	310	10
mature forest	0	-	-	-	-
disturbed forest	19	1	5.3	1	1.0
disturbed open	390	30	7.7	309	10.3

Table 9. Host-parasite data for all species of larval trombiculids found on reptiles and amphibians, 1980-1981.

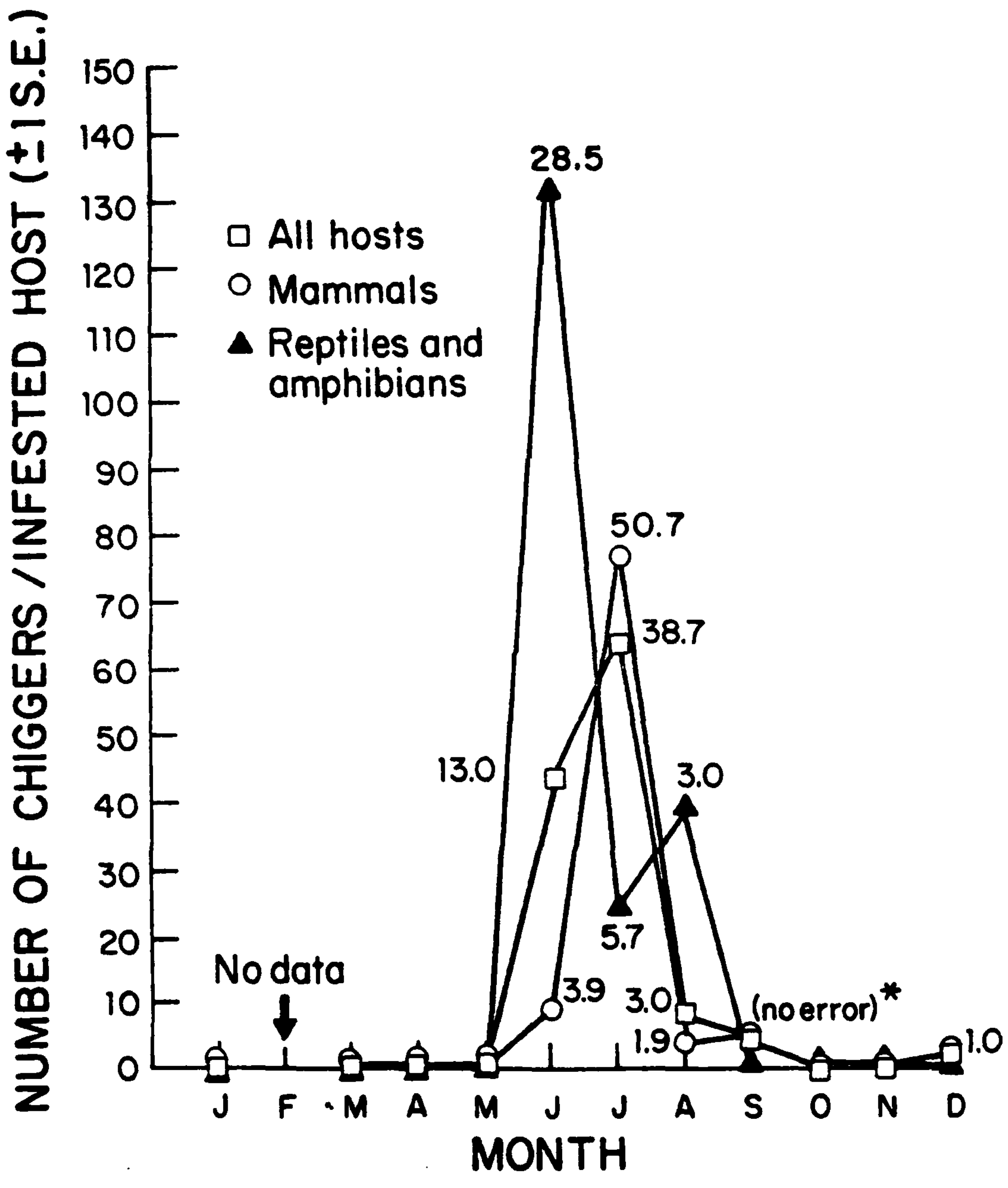
	number of hosts examined	number of hosts infested	percent of hosts infested	total number of chiggers removed	number of chiggers per infested host
Georgia	83	42	51.8	2210	67.0
Coastal Plain	14	12	78.6	830	69.2
Piedmont	69	30	43.5	1374	68.7
mature forest	1	0	-	-	-
disturbed forest	28	19	67.9	355	35.5
disturbed open	40	11	27.5	1019	101.9

behavioral niche differences explain these results. _S. hispidus_ is a ground-loving rodent of dense cover and meadows, and it tends to use established runs in moving about (Golley 1962, Hamilton and Whitaker 1979). _P. leucopus_ seldom makes and uses runs, is more inclined to semi-arboreal habits, and is more confined to thick brush and taller growth which allows it to reduce its contact with the substrate (King 1968). Its habits seem, in general, to make it less available to larval chiggers despite its more uniform distribution among habitat types (Table 7). While the low severities of infestation make it unlikely, it is possible that size differences between these rodents could explain some of the difference in infestation (Mohr 1961).

Reptiles and amphibians had highest rates and severities of infestation of all major taxa (Table 9). In the Piedmont, disturbed forest sites yielded the highest rate of infestation (67.9%), but disturbed open situations had the highest number of chiggers per infested host (101.9). Sites which were classified as "disturbed open" had often been logged and the ground strewn with slash and waste timber. In view of the close association between lizards (_Sceloporous_ and _Eumeces_), fallen logs, and pest chiggers, we find these results unsurprising.

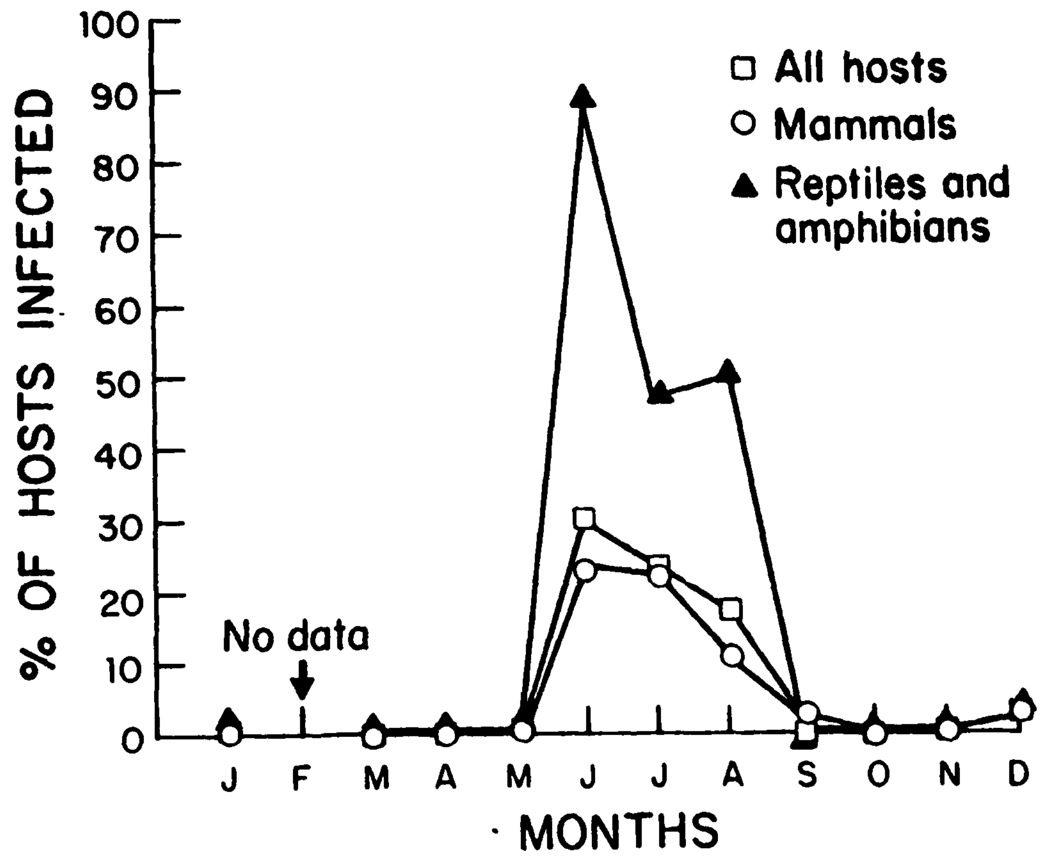
Analysis of seasonality of host-trombiculid relations are limited to data from the Piedmont with all habitats combined (Figures 2 and 3). The general pattern for both frequency and severity of infestation is one of a midsummer peak with a "tail" of recoveries into early winter. This pattern is typical of pest chiggers (_Eutrombicula_). To date, we have been relatively unsuccessful in accumulating quantitative information on "winter"

Figure 2. Total numbers of larval trombiculids (chiggers) removed from infested vertebrate hosts during each month, 1980-1981. Collections began in April 1980, and run through December 1981.



*one animal infested

Figure 3. Percent of vertebrate hosts infested by month, April 1980 - December 1981.


% OF HOSTS INFECTED
100
90
80
70
60
50
40
30
20
10
0
No data
All hosts
Mammals
Reptiles and amphibians
J
F
M
A
M
J
J
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S
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MONTHS

chiggers (some Trombicula, Euschoengastia and others, Loomis 1956). In February 1982, six unengorged E. peromysci were recovered from a single Peromyscus gossypinus from Jekyll Island, Glynn County, Georgia. In 1982, we hope to increase our understanding of trombiculids on the Coastal Plain (see following section). Reptiles show a peak in rate and severity of infestation in June. This is a month of egg laying for Sceloporous, Eumeces, and Terrapene (Mount 1975, Martof et al. 1980) and may be a time of enhanced association between reptiles and such preferred chigger habitats as old logs and loamy soils.

In the coming year, we propose to reduce our host survey effort in the Piedmont region and shift our attention to the Mountain and Coastal Plain areas. As we add to our data from these regions, we shall be able to determine whether the results and hypotheses we have formulated regarding the distribution of trombiculid mites and host organisms are generally applicable.

Our two year qualitative survey of hosts in the Piedmont has led us to formulate quantitative questions which we will address in 1982. The first question is: how is the density of unengorged larval chiggers related to density of potential host organisms? To answer this question, we will make quantitative estimates of vertebrate populations in limited areas. Reptile populations will be estimated by a mark-recapture system (Seber 1973), and small mammal populations will be estimated by removal trapping (Jensen 1975). Densities of larval chiggers will be estimated using "chigger samplers" and a modification of Menhinick's (1963) removal method (Wicht and Crossley 1982).

The second question we wish to answer is: how is severity of chigger infestation related to characteristics of the host organism? Size (Mohr 1961) and foraging habits (Easton 1975) have been shown to affect the number of chiggers per host. We will use a combination of field work (analyzing hosts by size within and among species and habits among species) and laboratory work (using established populations of larval chiggers) to attempt to answer this question.

A third question is: how is chigger infestation related to characteristics of the habitat? Again, a combination of field study (measuring environmental parameters *in situ*) and laboratory work (following infestation rate in a variety of experimental habitats) will be used to answer this question.

Finally, based on survey work in the Mountain and Piedmont regions, we hope by the end of 1982 to begin to quantify the factors controlling chigger distribution in these areas.

Literature Cited

Buckner, R. L. and L. N. Gleason. 1974. Arthropod ectoparasites and their seasonal occurrences on Microtus ochrogaster and Peromyscus leucopus from Warren County, Kentucky. Trans Kentucky Academy of Science 35:70-75.

Crossley, D. A., Jr. 1960. Comparative external morphology and taxonomy of nymphs of the trombiculidae (acarina). Univ. Kansas Sci. Bull. 40:135-321.

Crossley, D. A., Jr. and Charles W. Proctor, Jr. 1971. New records of chigger species (acarina: trombiculidae) in Georgia. Jour. Georgia Ent. Soc. 6:185-187.

Cunningham, J. R., S. E. Kunz and M. A. Price. 1977. Neoschongastia americana Development and longivity under laboratory and field conditions. Ann. Ent. Soc. Amer. 70:17-18.

Easton, E. R. 1975. Ectoparasites in two diverse habitats in western Oregon. II. Chiggers (Acari: Trombiculidae). J. Med. Ent. 12: 295-298.

Everett, R. E., M. A. Price and S. E. Kunz. 1973. Biology of the chigger Neoschongastia americana (Acarina: Trombiculidae). Ann. Ent. Soc. Amer. 66:429-435.

Gist, C. S., D. A. Crossley, Jr. and Virginia A. Merchant. 1974. An analysis of life tables for Sinella curviseta (Collembola). Env. Ent. 3:840-844.

Golley, F. B. 1962. Mammals of Georgia. University of Georgia Press, Athens.

Hamilton, W. J., Jr. and J. O. Whitaker, Jr. 1929. Mammals of the Eastern United States. Comstock Publishing Associates, Cornell Univ. Press, Ithaca, N.Y.

Huber, I. 1978. Prey attraction and immobilization by allomone from nymphs of Womersia strandtmanni (Acarina: Trombiculidae). Acarologia. 20:112-115.

Hubert, A. A. and H. J. Baker. 1963. The persistence of foci of Leptotrombidium akamushi along a transect in Malaya. Am. J. Hyg. 78:143-149.

Jayewickreme, S. H. and W. J. Niles. 1946. Successful feeding experiments with an adult trombiculid mite (Order Acarina). Nature. 157:878.

Jayewickreme, S. H. 1947. Rearing of Trombicula acuscutellaris Walch. Nature. 160:578.

Jenkins, D. W. 1947. A laboratory method of rearing chiggers affecting man (Acarina: Trombiculidae). Ann. Ent. Soc. Amer. 40:56-68.

Jenkins, D. W. 1947. The ecology and laboratory rearing of chiggers affecting man in the United States. Ph.D. Dissertation, Ohio State University.

Jensen, T. S. 1975. Trappability of various functional groups of the forest rodents Clethrionomys glareolus and Apodemus flavicollis, and its application in density estimations. Oikos 26:196-204.

King, J. A. ed. 1968. Biology of Peromyscus (Rodentia). Special. Publ. #2. American Soc. Mammologists. 593 pp.

Kulkarni, S. M. and P. V. M. Mahadev. 1973. Successful colonization of trombiculid mites with notes on life histories of three species. Indian J. Med. Res. 61:352-360.

Kunz, Sidney E. 1969. Biology and economic importance of the chigger Nedschoengastia americana on turkeys. Jour. Econ. Ent. 62:872-875.

Lawson, L. D. and R. W. Merritt. 1979. A modified Ladell apparatus for the extraction of wetland invertebrates. Can. Ent. 111:1389-1393.

Lipovsky, L. J. 1951. Collembola as food for chiggers (Acarina: Trombiculidae). J. Parasit. 37:324-326.

Lipovsky, L. J. 1954. Studies of the food habits of postlarval stages of chiggers (Acarina, Trombiculidae). Univ. Kansas Sci. Bull. 36:943-958.

Loomis, Richard E. 1956. The chigger mites of Kansas (Acarina, Trombiculidae). Univ. Kansas Sci. Bull. 37:1195-1143.

Martof, B. S., W. M. Palmer, J. R. Bailey, J. R. Harrison III and J. Dermid. 1980. Amphibians and reptiles of the Carolinas and Virginia. Univ. North Carolina Press, Chapel Hill.

Menhinick, E. F. 1963. Estimation of insect population density in herbaceous vegetation with emphasis on removal sweeping. Ecology 44:617-622.

Merchant, Virginia A. and D. A. Crossley, Jr. 1970. An inexpensive, high-efficiency tullgren extractor for soil microarthropods. Jour. Georgia Ent. Soc. 5:83-87.

Mohr, C. O. 1961. Relation of ectoparasite load to host size and standard range. J. Parasitol. 47:978-984.

Mount, R. H. 1975. The reptiles and amphibians of Alabama. Auburn Printing Co., Auburn, Ala.

Nadchatram, M. 1968. A technique for rearing trombiculid mites (Acarina) developed in a tropical laboratory. J. Med. Ent. 5:465-469.

Nadchatram, M. 1970. Correlation of habitat, environment and color of chiggers, and their potential significance in the epidemiology of scrub typus in Malay (Prostigmata: trombiculidae). J. Med. Ent. 7:131-144.

Nadchatram, M. and A. L. Dohaney. 1974. A pictorial key to the subfamilies, genera and subgenera of southeast Asian chiggers (Acarina, prostigmata, trombiculidae). Bull. Inst. Med. Res. Malaysia, 16:1-67.

Nutting, W. R. 1968. Host specificity in parasitic acarines. Acarolgia 10:165-180.

Ott, Lyman. 1977. An introduction to statistical methods and data analysis. Belmont, Calif.: Duxbury Press.

Seber, G. A. F. 1973. The estimation of animal abundance and reloted parameters. Griffin, London. 506 pp.

Simonova, V. 1977. Laboratory culturing of chiggers (Acarina: Trombiculidae). Folia Parasitologica (Praha) 24:285-288.

Srivastva, S. P. and B. L. Wattal 1973. Studies on the distribution of ectoparasites and their vertebrate hosts in urban, semi-urban peripheral, and sylvatic zones of Nagpur City and neighboring areas in Maharashtra, India. Indian Jour. Ent. 35:306-324.

Traub, R. and C. L. Wissemann, Jr. 1974. The ecology of chigger-borne rickettsiosis scrub typhus. J. Med. Ent. 11:237-303.

Whorton, G. W. 1957. Intraspecific variation in the parasitic Acarina. Syst. Zool. 6:24-28.

Wharton, G. W. and R. K. Carver. 1946. Food of nymphs and adults of *Neoschongastia indica* (Hirst 1915). Science. 104:76-77.

Wicht, M. C. and D. A. Crossley, Jr. 1982. Some observations and improvements in chigger collecting methods. Manuscript.

Wrenn, W. J. 1974. Notes on the ecology of chiggers from northern Michigan and the description of a new species of *Euchoengastia*. J. Kansas Ent. Soc. 47:227-238.

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